Advanced Diagnostic Approaches and Current Management of Internal Disorders of Select Species (Rodents, Sugar Gliders, Hedgehogs)

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KEYWORDS

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African pygmy hedgehogs (*Atelerix albiventris*) are unusual, but increasingly common pets. Due to strict regulations preventing importation of African pygmy hedgehogs into the United States, those in the pet trade are from captive bred populations. The European hedgehog (*Erinaceous europaeus*), although occasionally kept as a pet, is the most common British mammal seen in wildlife rescue and rehabilitation centers.[1] Both species are commonly kept as zoologic specimens.

Sugar gliders (*Petaurus breviceps*), marsupials native to Australia, Tasmania, New Guinea, and islands of Indonesia, are seen as pets in veterinary practices throughout the United States. These unique creatures are often presented for illness related to inadequate husbandry and/or nutrition. As clients become more educated about these animals, interest is being generated in finding increased levels of veterinary care.

Rodents such as rats (*Rattus norvegicus*), mice (*Mus musculus*), gerbils (*Meriones unguiculatus*), hamsters (*Mesocricetus auratus*), guinea pigs (*Cavia porcellus*), and chinchillas (*Chinchilla lanigera*) continue to be presented to veterinarians as pets and as collection species. There is a great deal of knowledge about these animals

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and their use as laboratory research species, but less is known about diagnosing and treating them as individual pets.

As their popularity increases, more and more practitioners are being asked to examine, diagnose, and treat these animals for a bevy of disorders and diseases. Many procedures and techniques used in both traditional small and large animal medicine are used for these species, with minor adaptations or considerations. This article examines available diagnostic tools and treatment methodologies for use in hedgehogs, sugar gliders, and selected rodents.

PHYSICAL EXAMINATION

Hedgehogs that are comfortable and accustomed to human contact can be examined with minimal restraint and the use of a clean towel or glove.[2] Physical examination in hedgehogs that are unaccustomed to handling or are nervous in an unusual situation can be a challenge. Therefore, an initial hands-off observation is a critical step to take note of any obvious abnormalities; this can often be accomplished by observing the hedgehog from a distance in a clear container.[3] The animal's carriage and ambulation status should be evaluated. When standing, hedgehogs should have a plantigrade stance with the body lifted off the substrate. There are several proposed techniques to encourage the hedgehog to not roll into a ball, including scruffing the hedgehog before it rolls into a ball, exposing it to water,[4] or placing it on a flat surface without handling for a few minutes.[5] However, these techniques even when safely performed rarely work well enough or long enough for a complete physical examination and assessment to be performed. Isoflurane anesthesia is often used to facilitate a thorough physical examination (**Fig. 1**). Particular attention should be paid to areas prone to disease in hedgehogs. Commonly seen diseases and disorders of pet hedgehogs include, but are not limited to, oral masses, periodontal disease, pneumonia, cardiomyopathy, trauma, and neoplasia.[3,5] Therefore, a thorough examination should include a complete examination of the oral cavity and mucous membranes, auscultation of heart and lung sounds, dermatologic assessment, abdominal palpation, visualization and assessment of the reproductive organs, and close inspection of the limbs and feet.

Fig. 1. An African hedgehog being induced with isoflurane anesthesia.

As with hedgehogs, sugar gliders are often best first examined with a hands-off approach to assist the clinician in evaluating for signs of respiratory distress, neurologic deficits, or evidence of trauma. Handling can be accomplished manually by holding the individual in cupped hands or while supporting the body, restraining the head at the mandibular joint.[6] Restraint by the tail is not recommended. The use of anesthesia is encouraged to perform a full physical examination.[7] Conditions seen in sugar gliders include, but are not limited to, problems associated with inadequate husbandry/nutrition, dental disease, and trauma. Therefore, complete visual examination and palpation of the sugar glider in addition to obtaining a weight, body condition score, temperature, pulse, and respiratory rate is appropriate.

Rodents differ greatly in their receptiveness to manual restraint and examination because of species differences, familiarity with being handled, and presence and severity of illness.[8] Rodents that are accustomed to handling may allow an examination without chemical restraint. However, even rodents accustomed to handling may become panicked due to the unfamiliar location and/or an underlying disease. Brief periods of gas anesthesia are often worth the risk in order to reduce stress for a physical examination. Review of individual species susceptibility to and prevalence of disease may help guide a thorough physical examination of the rodent. Observation of the rodent in the cage is an appropriate starting point for an examination, and allows evaluation of the respiratory rate and character, locomotion ability, and overall demeanor of the animal.[9] Rodents such as guinea pigs and chinchillas that are prone to dental disease may benefit from having the oral cavity examined at the end rather than the beginning of the examination.

Based on the history, initial observation, and physical examination, a problem-oriented approach may be used to define identified problems, determine appropriate diagnostic tests, establish treatment, educate clients, and monitor patient progress.[10] Diagnostic testing should be aimed at proving likely items or disproving unlikely items on a differential diagnostic list. Published research on diagnostic testing with information regarding test accuracy for the appropriate species should be used when available. However, many problem-specific diagnostic tests are not validated in exotic species. The use of a clinically healthy animal as a comparison can provide data that, although not statistically significant, can still aid in developing an index of support for a particular diagnosis. The diagnostician should keep in mind that many examinations and tests will need to be performed with the use of anesthesia.[11] Therefore, planning ahead to minimize the number of times an animal is subjected to anesthetic episodes is advised.

DIAGNOSTIC PROCEDURES

Venipuncture

A safe sample volume of blood in small exotic mammals is often estimated to be 1% of lean body mass.[12] If the animal sampled is geriatric, suspected to be anemic, hypoproteinemic, or otherwise compromised, smaller amounts of blood should be collected. Continued loss of blood through hematoma formation also needs to be considered. Limiting the blood collected to 0.5% of the animal's body weight is appropriate in ill or otherwise debilitated animals,[13,14] Techniques for collecting blood are often similar to those used in cats and dogs because the vascular anatomy is often comparable.[12] The challenge often stems from the small size of the vessels and the change in approach that is often required to access different vessels.

Obtaining blood from hedgehogs, sugar gliders, or rodents without the use of chemical restraint or anesthesia is typically only possible if the animal is severely

debilitated.[15,16] Isoflurane is frequently used for anesthesia. Small amounts of blood (even as small as one drop) are often worth the effort of collection.[11] In a hematocrit tube, blood volumes this minute can still provide indispensable information such as a blood glucose level, a blood smear for a differential, or a packed cell volume and total solids.

In hedgehogs, up to 0.5 mL of blood can be taken from the lateral saphenous and cephalic veins.[3,12] The lateral saphenous vein is often best sampled just below the stifle.[2] These vessels can be difficult to visualize and are prone to collapse.[12] Different techniques have been described to avoid the common complications of venous collapse and sample clotting.[5] These techniques include collecting the blood in a 25-gauge needle hub and then collecting the blood from the hub with a microhematocrit tube or by using a preheparinized tuberculin syringe outfitted with a 25- or 27-gauge needle. The femoral vein can be used for blood collection of up to 1 mL in hedgehogs. However, due to its anatomic location, care must be taken to not lacerate the closely approximated femoral artery.

Unlike hedgehogs, sugar gliders have cephalic, lateral saphenous, and femoral veins that are typically easily visualized.[7] As in hedgehogs these tiny vessels are easily collapsed, and the use of an insulin syringe with a 27-gauge needle is recommended for collection. These animals also have a ventral tail vein; warming the animal is recommended to aid in vasodilatation of this vessel.[12] A maximum of 0.1 to 0.25 mL may be obtained from each of the previously described venipuncture sites. Sugar gliders also have a medial tibial artery, which is readily visualized and from which 0.5 mL of blood may be collected. However, this vessel is very mobile and tends to roll, making successful bleeding difficult. Digital pressure should be applied immediately after drawing from the artery to aid in avoiding the formation of a hematoma.[6]

Small amounts of blood may be obtained from most rodents seen in clinical practice by use of the lateral saphenous veins.[12] In mice, venipuncture of the submandibular vein at the junction where it meets the jugular vein behind the mandibular joint has been described. Small amounts may also be collected from the cephalic vein of guinea pigs, hamsters, chinchillas, and other larger rodents. In almost all cases, the use of anesthesia to minimize stress and reduce complications associated with the blood draw is indicated.[13] Blood collection without the use of anesthesia from the dorsal tail vein of rats has been described.[17] Rats were preconditioned to handling and crawling in a towel with their tail gently extended for 2-minute periods 4 to 5 days before the actual collection. A small incision was made about 15 mm from the distal end of the tail and digital pressure was applied proximal to the incision while blood was collected in a microhematocrit tube. Venipuncture of the lateral tail veins of rats has also been described without the use of anesthesia.[12] However, venipuncture using manual restraint has been shown to interfere with a rat's ability to thermoregulate for up to 30 hours after sampling, and in venipuncture taking longer than 2 to 3 minutes, blood corticosterone levels were increased, indicating high levels of stress.

Complete blood cell counts, chemistry panels, and other more focused testing will require larger volumes of blood than can be gained from the techniques previously described.[11] The jugular vein is of sufficient size in hedgehogs to collect a diagnostic blood sample.[3] This vessel may be difficult to visualize, particularly in obese hedgehogs, but it runs between the ramus of the mandible and the point of the shoulder as in other small mammals.[12] To access the vessel, the hedgehog may be held in ventrodorsal recumbency with the legs stretched caudally, as described in ferrets. The authors have also had success with positioning the hedgehog in dorsoventral recumbency with the legs stretched caudally over the end of a table, as is typically

used to bleed the jugular of domestic cats and some ferrets. A 22- to 25-gauge needle on a 1- to 3-mL syringe may be used to collect this sample.

Approximately 0.5 to 1 mL of blood can be collected from the jugular vessel of a sugar glider.[12] The use of a 25- to 27-gauge needle on a 1-mL syringe is recommended for collection. Rodents, due to size and species variation, will differ somewhat with regard to the appropriate size of needle and syringe as well as the amount of blood that can successfully be drawn from the jugular.

A peripheral vessel not typically used in blood draws for cats and dogs, but used with some frequency in exotic small mammal practice, is the cranial vena cava. The internal and external jugular veins join into the cranial vena cava, which unites with the subclavian vein before it empties into the right atrium of the heart.[18] Venipuncture technique for accessing this vessel will differ slightly between species of varying size and anatomic diversity, but all will likely require the use of general anesthesia.[13] Possible risks associated with collecting a sample from the cranial vena cava include hemorrhage, hemothorax, penetration of the heart, and hemopericardium.

In species the size of a hedgehog, chinchilla, guinea pig, or rat, a 1- to 3-mL syringe and 25- to 27-gauge needle are appropriate for sampling.[12] In animals of smaller stature such as sugar gliders, mice, and hamsters, a 1-mL syringe with a 27-gauge needle or tuberculin syringe with a 30-gauge needle may be needed.[18] Individuals should be placed in ventrodorsal recumbency with the front limbs placed to the respective sides of the thoracic cavity.

Rats, mice, hamsters, and gerbils have well-developed clavicles that articulate between the sternum and the scapular-humeral junction.[18] Methods described for accessing the cranial vena cava of the rat are often also successful in hedgehogs.[12] A 0.5-inch length needle is inserted cranial to the clavicle and angled 45° toward the opposite leg.[14] Positioning the needle caudal to the clavicle and cranial to the first rib will force the needle into a more lateral position and makes successful venipuncture difficult.

In guinea pigs and chinchillas, the clavicle is not as well developed, and a needle (five-eighths-inch length) should be placed cranial to the manubrium and first rib.[14] Techniques used to successfully sample guinea pigs have been found to be successful in sugar gliders.[12] The needle should not be advanced more than 1 to 1.5 cm into the thoracic cavity (**Fig. 2**). If no blood is aspirated into the syringe when negative pressure is applied, the needle should be slightly retracted and redirected toward midline.

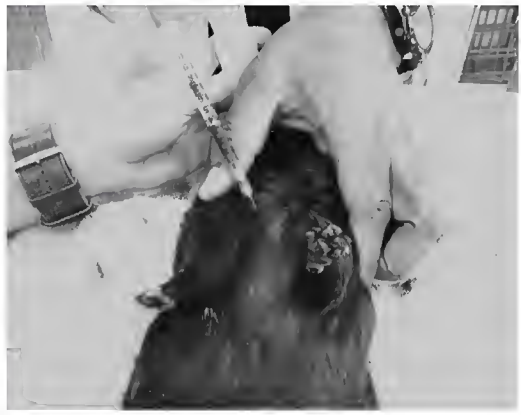

Fig. 2. Venipuncture of the cranial vena cava in a guinea pig.

Hematology and Immunoassay

More advanced hematologic testing, such as blood cultures, serology, antigen detection, and virus isolation, may be performed to further evaluate for evidence of disease.[16] Many diagnostic laboratories that assist with handling of laboratory animal specimens are also willing to run samples on individual pet animals, and most can run serology (immunofluorescent antibody [IFA] and enzyme-linked immunosorbent assay [ELISA]) on small amounts of blood (0.1 mL or less). Antigen detection via polymerase chain reaction (PCR) can also be performed on various samples including blood, nasal swabs, tracheal swabs, and fecal samples. Individual laboratories should be contacted in regard of their specific sample handling and shipping requirements. It is noteworthy that for some common microbes that infect small exotic mammal patients, such as *Mycoplasma* spp, culture results may be negative in infected animals.[19] In these animals, additional serologic testing such as ELISA or IFA in combination with PCR is often indicated. Providing the diagnostic agency with information regarding the disease process or processes you suspect may aid their recommendations for which tests or test packages to perform.

Many viral infections in small exotic mammals are asymptomatic unless further complicated via secondary bacterial infection.[20] Therefore, most diagnoses of viral infections in pets are based on clinical signs rather than serologic identification. Serologic testing for an individual is often not warranted because antibodies are often only present after clinical signs are abating. For households with multiple pets or that allow breeding, serologic testing to allow for disease prevention may be appropriate. Mice, rats, and guinea pigs are common laboratory species and, as such, microbial testing is readily available for these species. Hedgehogs and chinchillas have had reports of suspected infection with herpes simplex virus, but given their minimal representation as laboratory animals, additional hematologic testing is not routinely performed.[5] Gerbils and sugar gliders have no current reports of naturally occurring viral diseases and therefore routine serologic testing is also not commonly performed.[20] Serologic testing is available for select fungal[21] and parasitic organisms, and should be pursued when appropriate. For instance, sugar gliders have been shown to be highly susceptible to toxoplasmosis, and testing via ELISA or IFA may be indicated.[6,22]

Fecal Analysis

Fresh fecal samples can be evaluated via cytology and culture. Swabbing the rectum of small mammals gently with a culturette will supply a sample appropriate for fecal culture.[23] Anesthesia should be used for this procedure to decrease the likelihood of contamination of the sample and to decrease the risk of perforation. Suspected pathogens should be communicated to the diagnostic laboratory to aid in identification.[16] Some organisms, such as *Salmonella* spp, require different media for successful culture and are a potential zoonosis.

Fecal floatation and a direct smear are often sufficient in identifying most problematic parasites.[16] An anal tape test can also be employed to identify pin worms. Although rodents are often forthcoming with fecal samples, many species such as guinea pigs and chinchillas experience minor constipation within novel environments such as a veterinary hospital.[23] Hedgehogs also are often not forthcoming with fecal samples in a clinical setting.[2] Therefore, fresh fecal samples should be collected by owners whenever possible. If there is a delay between collection of the fecal sample and the transportation of the sample to the veterinarian, the sample should be kept refrigerated away from any human food items.

Urinalysis

Urinalysis can help identify urinary and reproductive tract disease. Collecting a free catch urine sample from a hedgehog may be attempted by placing the hedgehog in a clean plastic or stainless steel container.[5] Obtaining a sample appropriate for culture can be obtained via ventral percutaneous cystocentesis (ultrasound guided) or through sterile placement of a small flexible catheter.[2] Both procedures should be attempted only with appropriate anesthesia. Although urinalysis parameters have not been established for use in hedgehogs, neoplastic cells, crystals, white blood cells, and red blood cells may be considered abnormal findings depending on the collection method.[5] Urine test strips, although not verified in these species, can often be used successfully to help determine if a red hue observed in free catch urine is due to porphryin pigment or blood. A urine antigen test performed by MiraVista Diagnostics (Indianapolis, IN, USA) is also available for histoplasmosis, a fungal disease recently reported in an African pygmy hedgehog[24] and sugar gliders[11]; 2 mL of urine is requested by the company for antigen testing. However, analysis may be completed on a urine volume as small as 600 μL (Wheat LJ, MiraVista Diagnostics, Indianapolis, IN, personal communication, 2010). Multiple urine samples from one individual may be collected and refrigerated for up to 3 to 5 days to obtain sufficient sample size. However, the longer the delays in sample analysis, the higher the risk for contamination of other fungal growths that may cross-react and result in false positives. Other urine antigen tests are available for blastomycosis, aspergillosis, and coccidioidomycosis.

Cystocentesis can often be attempted in larger rodents such as guinea pigs, rats, and chinchillas, as described for the hedgehog.[23] Urine collection in sugar gliders and other common rodents can be difficult due to their small size. Sugar glider urine and fecal collection are further complicated by their unique anatomic composition. The urinary ducts, gastrointestinal tract, and genital ducts empty into a common cloaca.[25] Some species such as the gerbil are physiologically programmed to conserve water, and therefore only small amounts of highly concentrated urine are typically available.[23] These animals are often too small to safely perform cystocentesis or atraumatic urinary catheterization, and free catch samples should be collected when appropriate.

Cytologic Sampling

Additional and more advanced diagnostics are often pursued based on the clinician's assessment and localization of the disease process. Fluid in the thorax or abdomen can be collected via thoracocentesis or abdominocentesis (**Fig. 3**), respectively. The use of a 25-gauge butterfly catheter with a 3- or 6-mL syringe to perform thoracocentesis has been described in rodents.[14] Performing these procedures with ultrasound guidance is recommended. In addition to its potentially therapeutic value, the fluid removed can then be evaluated for protein levels, possible etiologic agents, cell population, and morphology. Samples should also be submitted for culture and sensitivity.

Although respiratory disease is often diagnosed on clinical suspicion and treated empirically, individuals that fail to respond to initial therapy often require additional diagnostics. The use of cytology with culture and sensitivity is often used to identify possible respiratory pathogens. Nasal swabs are used frequently and carry little risk to the individual, but these swabs are contaminated samples and do not accurately assess disease occurring in the lower airways. In stable patients, culture of samples collected via tracheal or bronchoalveolar lavage is recommended.[14] A method for endoscopic assisted tracheal aspiration in guinea pigs has been described.[26] The

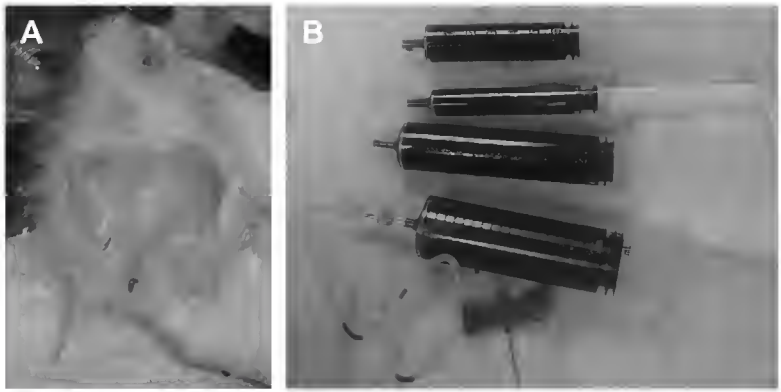

Fig. 3. (*A*) This hamster presented with severe abdominal distension. (*B*) Abdominocentesis was performed and cytology performed on the resulting fluid.

focus is on minimizing the fluid needed for sampling (approximately 0.5 mL), by positioning the guinea pig in sternal recumbency with the thoracic inlet lower than the remainder of the body.

Cytologic evaluation of the reproductive tract may also aid in diagnosis. Pouch infections have been documented with some frequency in captive female marsupials including the sugar glider.[25] The pouch should be swabbed and checked for yeast and bacteria.[27] Culture is recommended in addition to cytology. Males presenting with stranguria, hematuria, or penile trauma should be evaluated for prostatitis and involvement of the paracloacal glands common to both genders.[25] In hedgehogs, the predominant cause of abnormal vaginal discharge is reported to be neoplasia, but pyometra and metritis have also been reported.[3] Infection and inflammation of the uterus, testicles, vagina, epididymis, and mammary glands have been reported in rodents.[28] Cytologic evaluation with culture and sensitivity of any discharge or milk should be completed to determine etiology.[28]

Neoplasia is common in hedgehogs, sugar gliders, and rodents.[15] As a result, these animals are often presented for oncologic evaluation. Fine-needle aspiration, as in other species, is of minimal risk, but often the quantity and quality of cells evaluated is poor.[29] Aspiration is often completed with a 22- or 25-gauge needle for most sites and ultrasound guidance is used when appropriate. Biopsies, both incisional and excisional, are options that increase the likelihood of diagnosis (**Fig. 4**). Samples can be evaluated via cytology and histopathology. Additional stains and immunohistochemistry may also be completed depending on the index of suspicion for a particular diagnosis.

Evaluation of the bone marrow may be completed if neoplasia is suspected or for malignancy staging.[13] Other indications for bone marrow examination include hematologic disorders, anemia, thrombocytopenia, gammopathies, and lymphoproliferative disorders. The most common site for bone marrow aspiration and biopsy in small exotic mammals is the proximal femur. Other locations that may be used are dependent on the size of the patient but include the proximal tibia, proximal humerus, and the ileum. General anesthesia and infiltration of the area to be sampled with a local anesthetic, such as lidocaine, is recommended. After surgical preparation of the site, a small skin incision should be made and a spinal needle with stylet advanced into the medullary cavity. Once the stylet is removed, negative pressure is applied via the syringe until blood is seen and marrow has been aspirated. The use of excessive

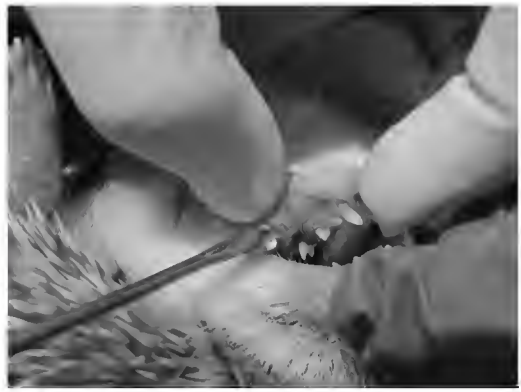

Fig. 4. Biopsy of a gingival mass in an African hedgehog.

negative pressure should be avoided because it can contaminate the sample with peripheral blood artifact, which hinders interpretation. Bone biopsy has been described with the use of an 18- and 20-gauge spinal needle.[29] Due to the small size of these pets, penetration of both cortices and exiting the skin through the opposite side may be attempted to preserve the core. The stylet can then be reintroduced before the needle is removed to push the sample out through the bottom of the needle (**Fig. 5**).

Radiographs

As with most of the previously described diagnostic techniques, taking diagnostic quality radiographs in all but the most severely ill hedgehogs, sugar gliders, and rodents will require the use of sedation and/or anesthesia.[30,31] Manual restraint may be attempted in animals that are critically ill and are at high anesthetic risk, but the stress induced from handling often limits its sole use. The use of anesthesia also lessens the likelihood that images will need to be repeated because it allows for correct patient positioning and greatly reduces the probability that the patient will move at an inopportune time.

Total body projections including the skull, extremities, and tail are the views taken most often for the species discussed here. Radiolucent tape is often used to

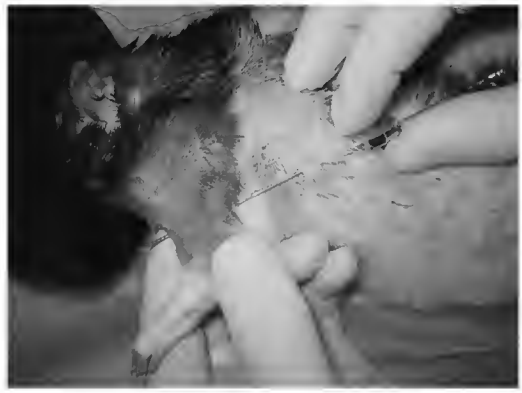

Fig. 5. Bone marrow biopsy of rat humerus using a 20-gauge spinal needle with stylet.

appropriately position limbs and anesthetic face masks when needed. Ventrodorsal and lateral projections are the most commonly taken views. When evaluating for diseases localized to the head including dental disease, multiple projections of the head including lateral, oblique, ventrodorsal, and rostrocaudal are needed.[32,33]

As with small animals, systematic evaluation of the thoracic and abdominal cavities is indicated. Comparing radiographs of patients with those taken of clinically unremarkable animals is one of the most beneficial ways to identify abnormalities (**Fig. 6**). Collections of radiographs featuring these species have been published demonstrating examples of both healthy and ill individuals.[15,30,31] Radiographs provide valuable, noninvasive supporting evidence for disease conditions commonly seen in these species including, but not limited to, respiratory disease, fractures, luxations, neoplasia, gastric distention, and impaction (**Fig. 7**). Radiographs are also beneficial in evaluating the urinary and reproductive tracts (**Fig. 8**).

Contrast Radiography

Contrast radiography may help identify and localize disease conditions of the gastrointestinal tract, urinary tract, and reproductive tract. The use of barium as a contrast agent has been described as a method to successfully evaluate gastrointestinal motility in the guinea pig.[34] Fluoroscopy may also be used for this purpose. Gastrointestinal-positive contrast studies with barium have been completed in multiple rodents including guinea pigs, rats, chinchillas, and hamsters.[31] In mammals, the uterus lies in close proximity to the colon, dorsal to the bladder, and when enlarged due to pregnancy or pyometra, can be difficult to distinguish from the colon (**Fig. 9**). A small amount of air or barium carefully injected into the colon can help differentiate between the terminal colon and reproductive tract. This technique is not recommended in sugar gliders because the gastrointestinal and urogenital systems have a common exit point.

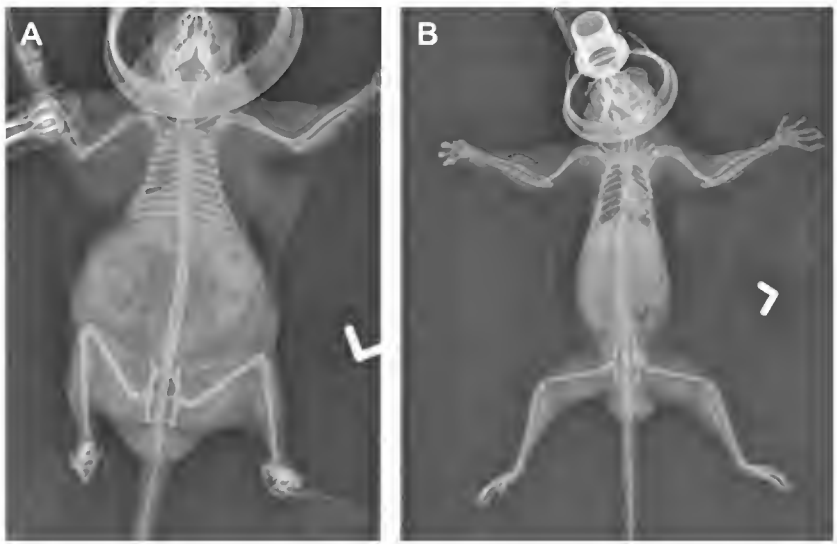

Fig. 6. Ventrodorsal radiographs of 2 sugar gliders. (*A*) Gastric and intestinal dilation is shown in this clinically ill animal. (*B*) A sugar glider with an unremarkable gastrointestinal tract, missing the distal phalanges of the first 2 digits of the right manus is offered for comparison.

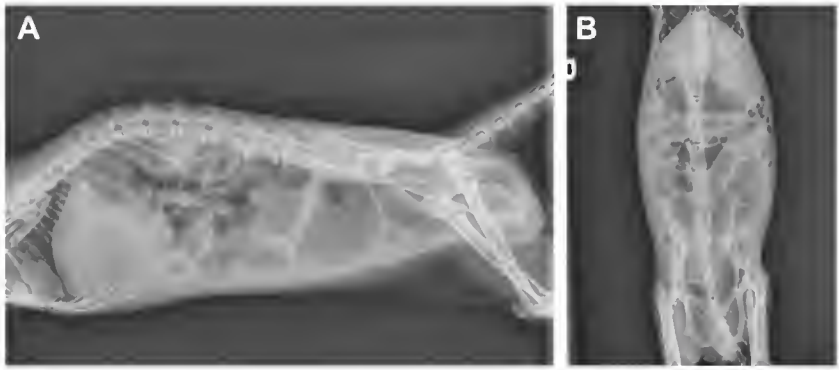

Fig. 7. Lateral (*A*) and ventrodorsal (*B*) radiographs of a chinchilla that show marked gas and fluid distention of the cecum as well as gas and fluid noted throughout the gastrointestinal structures.

Urinary tract disease is also common in these species and although plain radiographs are often sufficient to identify the presence and probable location of calculi, additional studies such as contrast cystography, urethrography, or intravenous pyelography may be indicated to evaluate for bladder wall integrity, concurrent disease conditions, and renal function.[35] In most cases, the small size of these patients makes these procedures technically difficult and time consuming. As a result, these diagnostics are rarely performed in private practice.

Another form of contrast radiography, myelography, is used in addition to survey films to more fully evaluate the spinal cord.[36] An example of successful myelography

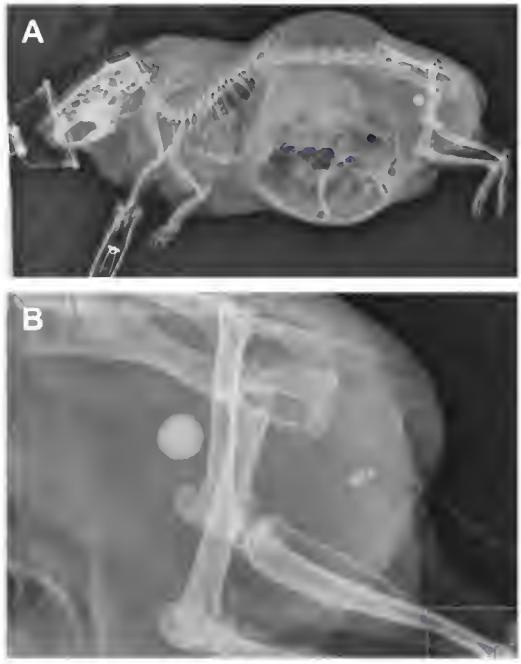

Fig. 8. (*A*) Right lateral radiograph of female guinea pig with urethral and urinary bladder stone. (*B*) Magnified image showing urethral and urinary bladder stone.

Fig. 9. A lateral radiograph of a female hedgehog 3 days postpartum with either dilated gastrointestinal loops or a gas-filled uterus. Additional diagnostics including contrast radiography or ultrasound are recommended.

in a clinically unremarkable guinea pig has been published.[31] Indications for completing a myelogram include trauma (luxation or fracture), suspect neoplasia, or other potential compression or space occupying lesion(s) affecting the brain or spinal cord.[36] Collection of cerebrospinal fluid to evaluate for inflammation, infection, or neoplasia may also be indicated and should be done before injection of contrast material. Although myelography and cerebrospinal fluid collection are relatively inexpensive, completing the procedures in extremely small patients can be technically challenging, and the potential disadvantages can include exacerbation of neurologic signs leading to seizures, paralysis, and death.

Most neurologic conditions in the sugar glider are attributed to trauma or inadequate nutrition.[7,11,15] In hedgehogs, neurologic signs have been attributed to multiple causes including trauma, toxins, infection, and malnutrition.[3,5] Wobbly hedgehog syndrome is a progressive neurologic disease that presents with ataxia followed by paresis and paralysis. This diagnosis is made either by clinical signs or via histologic evaluation of the spinal cord, brain, and peripheral nerves. Reports of clinical signs and pathologic changes consistent with intravertebral disc disease have also been described recently in African hedgehogs.[37] No reports of myelography in sugar gliders or hedgehogs are reported in the literature.

Magnetic Resonance Imaging

A less invasive, but more expensive method for imaging the neurologic system, including brain and spinal cord, is magnetic resonance imaging (MRI). In addition to providing clinical data to support neurologic diagnoses, MRI has also proven useful in evaluating other soft tissues within the thoracic and abdominal cavities. Despite its promising clinical use, there are no current reports of MRI used in a veterinary setting for diagnoses of pet rodents, sugar gliders, or hedgehogs. There is a report of MRI performed in Sprague-Dawley rats to determine appropriate techniques for evaluation of the rat brain and abdomen.[38] Multiple MRI studies have been completed in rodent models to research human disease conditions such as multiple sclerosis in mice[39] and pulmonary lesions from infection with *Mycobacterium tuberculosis* in guinea pigs.[40] Disadvantages of MRI usage in a clinical setting with these pets include lack of availability, cost, length of anesthesia time in animals prone to hypothermia, and lack of documented use in pets. It is hoped that as the breadth of usage of MRI in laboratory animal medicine is published, examples of clinically healthy species will be made more readily available to clinical practitioners focused on the care of pets.

Computed Tomography

Computed tomography (CT) has proven useful in evaluating the skull and vertebral column of exotic mammals.[36] Perhaps most promising is the use of CT as a modality for accessing rodents with dental disease.[41] CT enables the clinician to assess multiple slices of tissue, minimizing the effects of superimposition of bony and soft tissue structures seen in traditional radiographs. This modality allows for more thorough assessment of bone loss and osteomyelitis. CT is routinely used at the authors' facility for evaluating the extent of soft tissue and bony involvement of facial and/or tooth root abscesses (**Fig. 10**). In addition, the use of microCT has been reported to evaluate a tooth root abscess and osteomyelitis in a guinea pig.[42] MicroCT scanners offer superior resolution compared with conventional CT. However, microCT scanners are not readily available in clinical settings, aside from veterinary research facilities.

Ultrasound

Another imaging modality used with increasing frequency in small animal exotic practices and in the authors' facility is ultrasound. In many cases ultrasound is used instead of contrast procedures. Indications for abdominal ultrasound include palpable masses, fluid, or systemic disease to which a cause cannot be readily identified. Ultrasound provides greater imaging detail than that of contrast procedures for abdominal neoplasms and urogenital abnormalities.[14] Ovarian cysts (**Fig. 11**), dystocias, reproductive tract neoplasias, and urinary tract calculi are well visualized via ultrasound.[43] Pleural effusion, pulmonary pathology, and mediastinal masses may also be evaluated via thoracic ultrasound.[44] Cardiac structure and function may be examined via ultrasound in rodents.[45]

Endoscopy

Endoscopy has multiple uses with exotic mammal and marsupial medicine, but actual use is dependent on clinical signs and limited to some extent by the size of the patient.[46] The most common uses for endoscopy in these small patients are assisted tracheal intubation and oral examination. The ability to appropriately diagnosis and

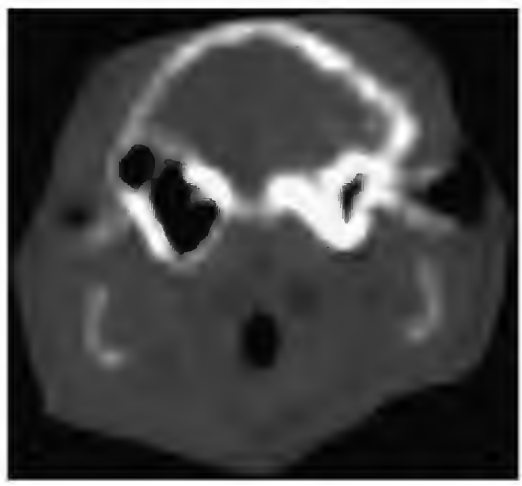

Fig. 10. A computed tomographic image of a guinea pig with chronic severe left-sided otitis media/interna and osteomyelitis.

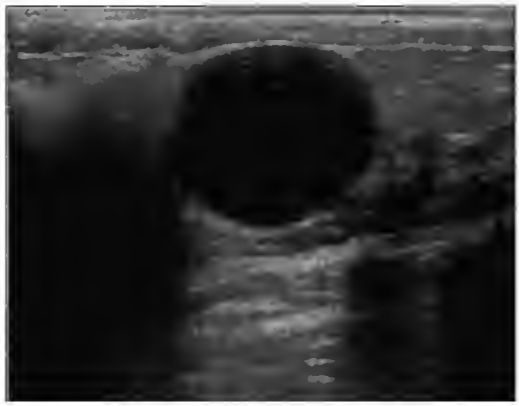

Fig. 11. Ultrasound of a guinea pig showing polycystic ovarian disease.

determine the extent of dental disease and associated pathology is greatly enhanced by endoscopy because of focal illumination and magnification. The use of the endoscope may also aid in biopsies and foreign body retrieval, and serve as a surgical aid in the oral and paranasal cavities. The use of endoscopy to identify and take samples of pathology of the distal urogenital tract has also been described for use in larger rodents such as guinea pigs.[47]

Necropsy

A final and very valuable diagnostic method is necropsy. Although every effort is made to prolong a high quality of life, individuals that have succumbed to disease present a unique opportunity to learn valuable information. Much of our understanding in these species has been gained from necropsy reports that have given us valuable information regarding commonality and pathology of certain disease processes. As we learn more about the disorders and diseases that incapacitate these animals, our ability to accurately diagnose and identify appropriate treatments continues to improve. The pairing of this knowledge with the ever increasing medical technological advances promises great improvements for the practice of exotic veterinary medicine.

THERAPEUTICS

Although many dosages and delivery methods currently used in these animals are anecdotal, clinical improvement is noted in many individuals. Multiple resources have published data on drug usage in hedgehogs and sugar gliders.[15,16,48] However, most of the information lacks pharmacokinetic and pharmacodynamic backing. Pharmaceutical research is more readily available for rodent species commonly kept as laboratory animals. However, much work remains for determination of pharmaceuticals appropriate for rodents kept as pets.

Subcutaneous administration of medication and fluids is typically successfully delivered in the hedgehog.[5] However, due to the large amount of adipose tissue present in the subcutaneous layer, there may be decreased or delayed uptake or distribution. Subcutaneous medications are also easily administered in sugar gliders and most rodents.[11,14] Intravenous medications can be difficult in these small animals, and venous catheters are dislodged when hedgehogs roll into a defensive position. Intraosseous catheters are often preferred in critically ill patients when fluids, colloids, or intravenous antibiotics are indicated. Oral medications work well in patients that are

Fig. 12. Oral antibiotic administration in a young hoglet.

stable and interested in food (**Fig. 12**). Many veterinarians advocate mixing oral medication with small amounts of juice or a favorite food such as banana or peanut butter, and report clinical improvement using this method.[5] However, studies are lacking that identify what effect, if any, mixing the various drugs with foods and liquids of varying pH and chemical composition has on the activity and bioavailability of different drugs.

Therapeutics should be based on results of a thorough diagnostic evaluation. In more critically ill animals or those with significant financial limitations, empirical supportive care may be initiated with regard to background information about the species being treated and the clinical signs of the individual animal.[10] Despite some of the current challenges in diagnosing and treating pet sugar gliders, hedgehogs, and rodents, increasing experience with examining and caring for these animals is furthering veterinarians' ability to provide quality medical care to these beloved and special pets.

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